SAMPLING AND ANALYSIS PLAN

Kaelepulu Stream System
Oahu, Hawaii

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Revised November 2009

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Region 9 Quality Assurance Manager

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1.0 INTRODUCTION

The purpose of the sampling effort is to produce data for development of Total Maximum Daily Loads (TMDLs) for the Kaelepulu Stream Watershed system which consists of Kaelepulu Estuary, Kaelepulu Stream and Hamakua Stream. These waterbodies are listed as impaired by the Hawaii State Department of Health, Environmental Planning Office (DOH) for Section 303(d) of the 1972 Clean Water Act (CWA). The waters do not meet water quality standards for nitrogen, phosphorus, sediment, enterococci, and chlorophyll a. The sampling effort will determine the actual extent of non-compliance and specifically provide water quality and hydrologic/meteorologic data necessary to calibrate a watershed model needed to establish the TMDLs. The effort will also include a sanitary survey.

1.1 Site Name or Sampling Area

Kaelepulu Watershed

1.2 Site or Sampling Area Location

The Kaelepulu Stream Watershed is located on the windward side of the Island of Oahu in the Kailua neighborhood, State of Hawaii. This area is commonly known (by the residents) as Enchanted Lake; the Hawaiian name for the area is Kaelepulu.

1.3 Responsible Agency

Project completion will be assisted by the incorporation of a principal investigators from the College of Tropical Agriculture and Human Resources, Department of Molecular Biosciences and Bioengineering (MBBE) to oversee completion of the proposed project objectives and an agreement with the University of Hawaii Water Resources Research Center for laboratory analytical services. The Project Organization are summarized in the following table.

1.4 Project Organization

Title/Responsibility	Name	Phone Number
EPA Project Manager		
Project Manager	**David Penn**	(808) 586-4339
Staff	**Renee Kinchla**	(808)-586-4369
Quality Assurance Manager		
Contractor (Company Name)	**University of Hawaii**	
Contractor Staff	**Dr. Clyde Tamaru**	808-342-10963
	Dr. Roger Babcock	(808) 956-7298

1.5 Statement of the Specific Problem

Section 303(d) of the 1972 Clean Water Act (CWA) directs each State to develop a list of water bodies that do not meet State water quality standards. In Hawaii, the Department of Health, Environmental Planning Office (DOH) has been tasked with this responsibility. Water bodies that are so listed do not meet water quality standards for specific constituents and thereby inhibit beneficial uses of the water body, such as for recreational uses or for supporting fresh water or marine wildlife. In 1998, DOH issued its initial list of impaired water bodies. This list was updated in 2001 and again in 2004. The 2004 list includes Kaelepulu Stream and three receiving areas that may be impacted by the stream – Lanikai Beach Station, Kailua Beach Station and Oneawa Beach Station. The specific problem addressed herein is the development of TMDLs

for the Kaelepulu Stream Watershed system which consists of Kaelepulu estuary, Kaelepulu stream and Hamakua Stream. The system is water quality limited in terms of nitrogen, phosphorus, sediment, enterococci, and chlorophyll a.

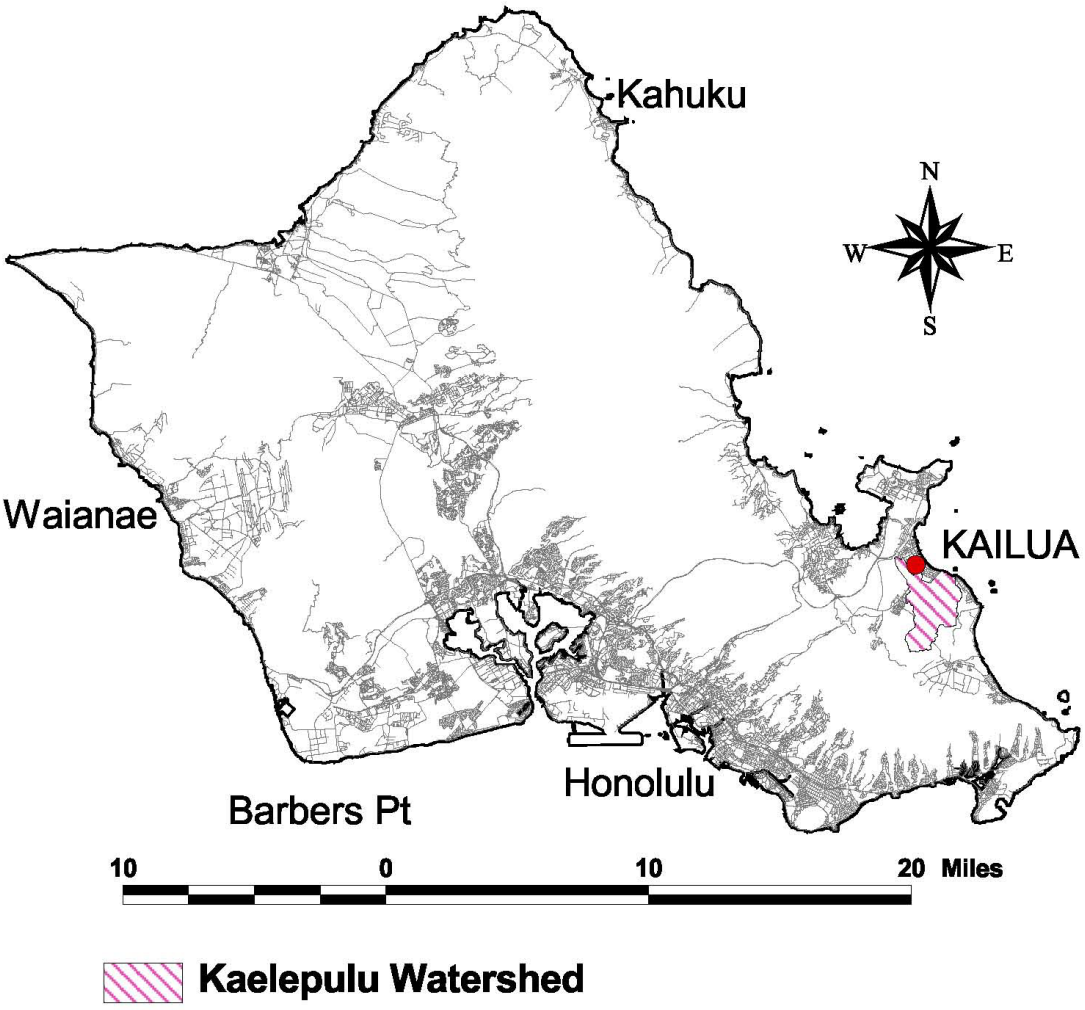
2.0 BACKGROUND

The purpose of the sampling effort is to produce data for development of TMDLs for the Kaelepulu Stream Watershed system which consists of Kaelepulu estuary, Kaelepulu Stream and Hamakua Stream. The system is water quality limited in terms of nitrogen, phosphorus, sediment, enterococci, and chlorophyll a. The Kaelepulu Stream Watershed is located on the windward side of the Island of Oahu in the Kailua neighborhood, State of Hawaii (see Figure 2.1). The sampling effort will also shed light on the existing degree of impairment which is not well established and will include a sanitary survey.

2.1 Site or Sampling Area Description

The site or sampling area (watershed) occupies 3,450 acres of mixed uses including residential (2,043 acres), preservation (1,122 acres), agricultural (275 acres), and industrial (12 acres) zoned areas. The site or sampling area is bordered on the north by Kailua Town, on the west by Kawainui Marsh and Maunawili, on the south by Waimanalo, and on the east by Lanikai and the Pacific Ocean. The specific location of the site or sampling area is shown in Figure 2.2.

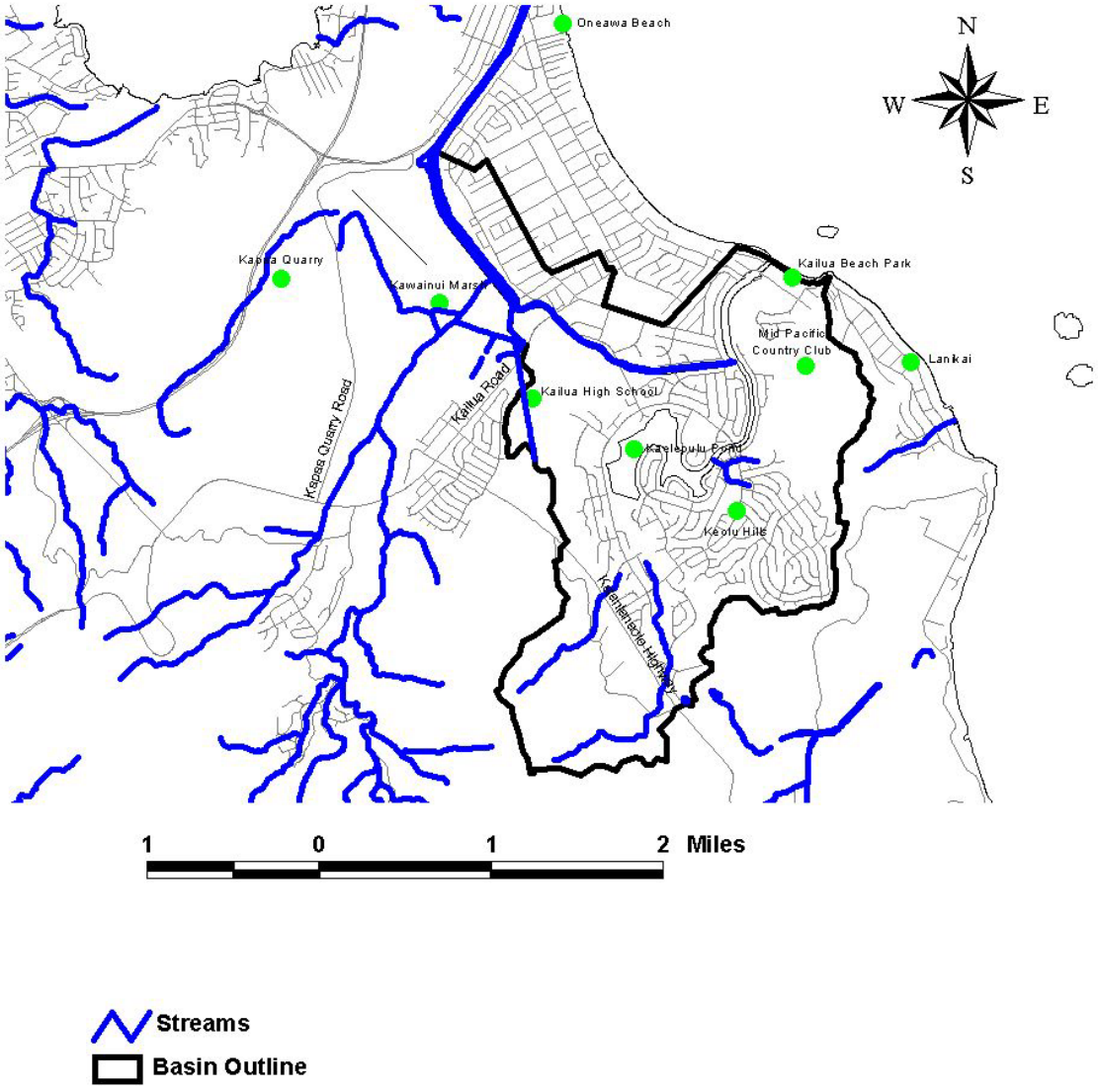
The Kaelepulu Estuary, also known as Enchanted Lake, is a remnant of an estuary that contained an ancient Hawaiian fishpond that had perhaps three times the surface area of the existing ponded estuary segment. The ponded estuary segment was dredged by developers beginning in the 1940's to a reported depth of 10 feet. Over the ensuing 65 years, numerous developments have sprung up in the drainage area and have contributed significant sediment and other pollutants. The watershed today is largely although not completely built-out. Throughout this period of development, the land usage has been mostly residential (2/3) and preservation (1/3). Therefore, the pollutant loadings have been typical of residential urban runoff and preservation land runoff. The watershed contains, homes, streets, schools, churches, parks, a golf course, wetlands, forested preservation lands, light industrial/shopping areas, gas stations, some cesspools and septic tanks.



Kaelepulu Sampling and Analysis Plan

Figure 2-1. Location Map

November 2005



Kaelepulu Sampling and Analysis Plan

Figure 2-2. Site Map

November 2005

2.2 Operational History

The Kaelepulu Watershed Stream system waterbodies have both point and non-point pollutant sources/inputs. Hamakua Stream has non-point source inputs from the Hamakua Wetlands and point source inputs from adjacent residential areas (stormdrains). Kaelepulu Estuary has point source inputs from adjacent residential areas, schools, parks, a golf course, commercial areas, and a state highway (stormdrains), and non-point source inputs from wetlands and forested hillside preservation lands. Normal beneficial use of these zoned land uses create some polluted runoff containing sediment, nutrients, and bacteria.

2.3 Previous Investigations/Regulatory Involvement

The previous sampling efforts are described in detail in the separate document: Kaelepulu Stream System TMDL Scoping Report.

2.4 Geological Information

No data currently exist for groundwater in the study area. No drinking water supply wells are currently located in the study area and no other wells are known to exist. The subsurface in the area is a well-developed dike complex (volcanic structures containing vertically oriented impervious formations lined up in parallel interspersed with more pervious material from a prehistoric caldera) overlain with alluvial sediment. This geologic formation is not a candidate for drinking water supply due to low potential for substantial sustainable yield. There is no known groundwater flow direction at the site. The Honolulu Board of Water Supply (BWS) chief geologist (C. Lao) has stated that there is little chance that groundwater has any significant impact on the Kaelepulu Watershed system.

2.5 Environmental and/or Human Impact

The pollutants of concern include nutrients (nitrogen and phosphorus), sediment, bacterial contamination (enterococci) and chlorophyll a. The possible and actual impacts of the nutrients include biostimulation of bacteria and algae which can reduce water clarity, possibly reduce or

eliminate dissolved oxygen causing odors and fish kills under extreme conditions. The sediment impacts water clarity, water circulation due to changes in bottom profiles, flora and fauna populations, and indirectly bacteria counts due to association of indicator organisms with the particulates. Bacteria contamination if due to human sewage can cause disease transmission to humans who contact the water bodies or eat fish etcetera. Because Kaelepulu Stream empties into Kailua Bay at Kailua Beach which is the most popular beach on Windward Oahu, there is potential for adverse impacts to human health due to sewage contamination. Bacteria contamination can also cause disease in fish and other exposed fauna. Chlorophyll a is a measure of algal concentration and the impacts of excessive concentrations include reduced water clarity, possible odors, and possible algal toxicity that can affect fish and/or humans in extreme cases.

3.0 PROJECT DATA QUALITY OBJECTIVES

3.1 Project Task and Problem Definition

The purpose of the sampling effort is to produce data for development of TMDLs for the Kaelepulu Stream Watershed system. The system is water quality limited in terms of nitrogen, phosphorus, sediment, enterococci, and chlorophyll a. In order to determine TMDLs, a model of the system must first be created to accurately simulate its response to pollutant inputs. The developed model is then used to predict the maximum amount of pollutants that can be input to the system while still achieving the established Water Quality Standards. The sampling effort will also shed light on the existing degree of impairment which is not well established and will also include a sanitary survey. Soil and sediment sampling are not included in the sampling effort.

The system model must simulate circulation/hydraulics, sedimentation, chemical and biochemical reactions within the various waterbodies. Inputs will include surface inflows/outflows, groundwater, ocean tides, sedimentation, etcetera. TMDLs may be appropriate for several different conditions such as average annual, dry period (May 1 – October 31), wet period (November 1 – April 30), dry period storm event, and wet period storm event. TMDLs may not be needed for all of these scenarios. In any case, the type and complexity of the chosen modeling approach will dictate sampling needs.

A moderate complexity model would be essentially one-dimensional and treat the Kaelepulu estuary as a well-mixed single volume of water that empties into Kaelepulu Stream which is joined by Hamakua Stream on its way to its terminus at Kailua Beach. The data requirements would include good outflow data, weather data, storm drain input hydrographs, stream hydrographs, concentrations of pollutant inputs from sub-basins and permitted dischargers, groundwater inputs, and lake level data. Such a model would not be able to predict the pollutant concentrations at different locations in the estuary, but it would be able to predict the average concentration of each pollutant in the outlet of the estuary and along the lengths of the streams. The required data for input to such a model would be significant, but less than a high complexity model.

A high complexity model would require the most data to calibrate and validate. It would also require the most time and effort to create and implement. Such a model would be a 3-dimensional dynamic circulation model that would consider the tides, seawater-induced density gradients, temperature gradients, convection and dispersion, biological action, deposition and nutrient loss, resuspension, groundwater inputs, bathimetry, and estuary level data. In order to calibrate such a model, extensive time-series data on density and temperature gradients as well as flow fields correlated to tides, and pollutant concentration profiles would be needed throughout the system. The benefits of the complex approach include the possibility that the model will highly accurately simulate the system allowing assignment of waste loading allocations at a very fine level, that it will accurately establish the extent of existing impairment and allow highly targeted and effective clean-up and control/management practices. The main drawback of the complex approach is the cost associated with collecting the massive amount of data that would be required for such a model.

Any model should also be able to simulate how the watershed will behave following mechanical dredging at various locations to remove sediment. There are certain areas containing significant sediment buildup for which dredging plans have been proposed.

Several models have been briefly reviewed for appropriateness. These included several surface water models distributed by the EPA Center for Exposure Assessment Modeling (CEAM)(www.epa.gov/ceampubl/swater/index.htm) including EXAMS, HSCTM2D, HSPF, QUAL2EU, SED3D, SWMM, and WASP. Several of these models as well as others could be appropriate for the Kaelepulu Watershed. EXAMS does not seem to be geared to watersheds. HSCTM2D seems to be a detailed 2-D model for sediment transport, deposition, resuspension, dispersion, aggregation and pollutant adsorption and desorption but not geared toward nutrient chemical transformations which will be important for Kaelepulu estuary. HSPF seems to be quite powerful in that it is able to model all of the relevant inputs, the nutrient chemistry, etc., however, it is apparently limited to well-mixed rivers and reservoirs. HSPF could be a good choice if it is desired to model the estuary as a well-mixed reservoir. QUAL2EU is an interesting model for branching stream systems. It is not completely clear how it would handle a lake, but apparently as a well-mixed detention basin (similar to HSPF). QUAL2EU seems better suited than HSPF to simulating nutrient cycles, chlorophyll concentration, and bacteria transport. SED3D is a complex 3-dimensional sediment transport model that can incorporate tides, freshwater inflows, density gradients, complex bathimetry and shoreline geometry. This model

is not geared toward simulating nutrient cycles or bacteria transport. SWMM is an urban runoff model that is good for simulating storm sewer systems and rivers using storm hydrographs. It can simulate aquatic biology, sediment transport, etc. SWMM would likely treat the lake as a well-mixed basin. WASP seems to be a good model for 1, 2, or 3-D simulation of water quality including pathogens and aquatic biology/nutrients. It apparently must be linked to a separate hydrodynamic/sediment transport model.

It is apparent that there are several possible choices of numerical model to simulate the Kaelepulu system. It will be most appropriate for the TMDL development contractor to make the final model selection based upon further review, existing experience and expertise. The following points are important:

1. The field sampling program will determine the spatial variability of water quality in the Kaelepulu estuary and streams. And, the spatial variability will determine how accurate or inaccurate it will be to treat the estuary as well-mixed in a simulation model.
2. If the spatial variability is low and the estuary can accurately be simulated as well-mixed, then any of several models can be selected based upon contractor preference. In rank order, probably QUAL2EU, followed by HSPF, followed by WASP linked to SWMM would be good choices.
3. The water quality standards for base flow conditions (non-rainfall events) are geometric mean based. If the standards are interpreted as the geometric mean of all water quality values from all sample locations in the estuary (or stream) taken in a given season (wet or dry or annual), then it may be perfectly appropriate to consider the system as well mixed (assuming there are an adequate number of sampling locations to give statistical significance).
4. If spatial variability is high and water quality should be simulated at different locations in the lake, then a more complicated model could be justified. In this case, additional time and funding will be required in order to develop, calibrate, and implement such a model.
5. For the purposes of this SAP, it will be assumed that data are needed to satisfy only a medium complexity model. The recommended conceptual approach will be to model the estuary as a well-mixed detention basin. Intuitively, this approach is satisfying because visual observation as well as water quality monitoring data suggest that the estuary is a collector of sediment and other pollutants. This means that flow fields, density gradients, and bathymetry at each sampling location will not be required.

The five steps that should be followed to develop TMDLs for the Kaelepulu Watershed system are as follows:

1. Develop a conceptual hydrodynamic and physical/biological model of the watershed system.
2. Find a suitable mathematical model (or models) to simulate the watershed system including all sources/inputs and their reactions/fate in the system that affect the limiting water quality parameters. The system will require a lake model component to simulate the estuary, an urban runoff component to simulate urban runoff including bacteria inputs, and a component to simulate the flows in the stormdrains as a function of precipitation. The model must also handle the theoretical effects of improved circulation following potential dredging operations.
3. Conduct sampling plan to calibrate the simulation model such that it can predict the observed water quality data set. This includes sampling of water quality in the system as well as in all inputs. The sampling must also include a sanitary survey.
4. Use the model to determine the maximum daily loads (TMDLs) that the watershed system can handle a still meet the water quality standards. Several scenarios could be investigated such as gradually reducing each known input until the model shows that the standards are achieved. The inputs could be reduced from each source by equal amounts or by equal percentage reductions, or other method such as a subjective analysis of "getting the low hanging fruit" or considering "best bang for the buck" control approaches.
5. Determine waste load allocations (WLAs), load allocations (LAs), and a safety factor to estimate input reductions necessary to achieve water quality standards. Assign the WLAs to permit holders.

Of these five steps, this SAP is directly solely at achieving step (3).

3.2 Data Quality Objectives (DQOs)

The 7-step DQO process as documented in EPA QA/G-4 "Guidance for the Data Quality Objectives Process" (EPA/600/R-96/005, August 2000) was followed to develop Data Quality Objectives for this study. Step 1 of the DQO process involves "Stating the Problem" (establishing the planning team including decision makers, defining the problem and developing a conceptual model of the hazard, and identifying available resources, constraints, and deadlines). The planning team consists of Dr. Roger Babcock, Greg Arakaki, June Nakamura, David Penn of the Hawaii State Department of Health (DOH), and the Kaelepulu TMDL workgroup which consists of various community stakeholders (Appendix A). The decision maker is the State DOH and the EPA Region IX. The problem is described as determination of existing water quality non-attainment in the Kaelepulu Watershed system (nutrients, sediment, bacteria, and chlorophyll a), determination of allowable maximum discharges of these contaminants into the watershed system such that established water quality standards are achieved (TMDLs), and allocation of allowable loads to specific source categories and permit holders in the watershed. The conceptual model consists of a watershed system containing several land uses (predominantly suburban), surface water bodies (Kaelepulu estuary, Kaelepulu Stream, and Hamakua Stream), an underlying groundwater system which may or may not interact with surface waters, wetlands, stormwater runoff from roads, parks, schools, residences, conservation lands, and other land uses, and other discharges from various sources (including birds and other animals, septic tanks and cesspools, etc.). The surface waters drain into the ocean at Kailua Beach which is a very popular internationally-ranked beach and where swimmers could contact any released contaminants. The surface water system behaves like a batch system in which the outlet to the ocean is mechanically opened roughly one day per month to allow drainage of water and contaminants and possible flushing of seawater back into the system. Generally, the estuary water level drops by up to 3 feet before the outlet at the beach closes off due to wave action (within hours to a few days). The available resources include approximately $255,000 from EPA to complete the TMDL process ($149,000) and sanitary survey ($56,000) for this watershed, sample analytical services provided by the Hawaii DOH Water Quality Lab, and a potential network of stakeholder volunteers for various data collection activities. The constraints include funding availability, and timeline issues such as the need to complete a TMDL draft technical report by April 1, 2006. The timeline was to begin sampling during the wet weather season November 1st – April 30th and collect samples for one year 2005-2006, and the funding deadline to complete the study is August 31, 2006 (Final Draft Technical Report). The deadlines for the draft and final draft reports were not met and were to be modified during a contract agreement with the University of Hawaii and forms the basis for the current

report. TMDL studies have been conducted on several stream systems on Oahu and each have used different approaches. Each of the existing TMDLs was somewhat simpler that the Kaelepulu watershed which is the first to involve an estuary and the first to involve bacterial contamination. Some lessons learned in the earlier TMDLs may be valuable in the current study include, 1) load allocations should be broken out by permit holder rather than by drainage basins, 2) there needs to be a good assessment of background concentrations of constituents of concern, 3) there needs to be an assessment of the extent of build-out in the watershed, 4) rainfall data assessment is important since it greatly affects hydrologic model output, 5) there needs to be an assessment of prior land uses as they pertain to water quality issues.

Step 2 of the DQO process involves "Identifying the Decision" (identify the principal study question, define alternative actions, develop a decision statement, and organize multiple decisions). The principal study question is: What are the Kaelepulu Watershed Stream system TMDLs for nitrogen, phosphorus, sediment, and bacteria such that water quality criteria are consistently achieved throughout the watershed? Study Question: Are water quality standards for nitrogen, phosphorus, sediment, bacteria, and chlorophyll a currently achieved throughout the Kaelepulu watershed? Alternative actions: a) do nothing, b) determine TMDLs/WLAs/LAs and then enforce reduction requirements through discharge permits. Other questions that are important include 1) What is the water quality variability within the Kaelepulu estuary and the Kaelepulu and Hamakua streams? 2) What are the existing and potential sources of bacteria contamination in the watershed (sanitary survey)?

A set of decision statements that need to be addressed in the SAP are as follows:

1. Do concentrations of nitrogen, phosphorus, sediment and bacteria in the Kaelepulu watershed meet applicable water quality standards? If yes, then do nothing. If no, then determine TMDLs.
2. Do sample results at a given location vary significantly on a seasonal basis (dry season versus wet season)? If yes, then develop wet weather and dry weather TMDLs. If no, then develop only one set of "annual" TMDLs.
3. Do sample results at a given location vary significantly on a monthly basis (within a single season)? If yes, then develop TMDLs on a practical worst case basis using the worst month data. If no, then develop TMDLs based on the average month data.
4. Do sample results at a given location vary significantly on a monthly cycle (within an single month due to a lack of ocean discharge)? If yes, then develop TMDLs on a practical worst case basis using the worst day data based upon a worst case month of

inputs. If no, then develop TMDLs based on an average day in an average month.

5. Are there enough sample results to calibrate the system simulation model? If yes, then sampling is complete. If not, then collect additional sampling such that sufficient data are available to accurately calibrate the system model.

Step 3 of the DQO process involves "Identifying the Inputs to the Decision" (identify the information needed, determine sources for the information, determine the basis for determining the action level, identify sampling and analysis methods that can meet the data requirements).

The water quality data needed to develop the TMDLs include:
Total nitrogen, detection limit less than 180 μg/L (about 100 = good)
Nitrate + Nitrite nitrogen, detection limit less than 8 μg/L (about 4 = good)
Ammonia nitrogen, detection limit less than 6 μg/L (about 3 = good)
Total phosphorus, detection limit less than 25 μg/L (about 10 = good)
Total suspended solids, detection limit less than 10 mg/L (about 5 = good)
Turbidity, detection limit less than 1.5 NTU (about 0.5 = good)
Chlorophyll a, detection limit less than 2 μg/L (about 1 = good)
Enterococcus, detection limit = 1/100mL
Analytical methods which can achieve these detection limits are given in Table 3.1.

Table 3.1 Study analytes and methods.

ANALYTE	UNITS	DETECTION LIMIT	METHOD
Total Nitrogen	µg/L	100	[1]Std. Mtd. 20th Ed., 4500-N_{org} D
Nitrate + Nitrite Nitrogen	µg/L	1	[2]Parsons et al, Method 1.1 + 1.2
Ammonia Nitrogen	µg/L	1	Parsons et al, Method 1.4
Total Phosphorus	µg/L	10	Std. Mtd. 20th Ed., 4500-P E
Total Suspended Solids	mg/L	0.1	Std. Mtd. 20th Ed., 2540 D
Turbidity	NTU	0.01	Std. Mtd. 20th Ed., 2130 B
Chlorphyll a	µg/L	0.2	Parsons et al, Method 4.1
Enterococcus	CFU/100mL	1	Std. Mtd. 20th Ed., 9230 C

[1] Standard Methods for the Examination of Water and Wastewater, 20th Edition, 1999.
[2] A Manual of Chemical and Biological Methods for Seawater Analysis, Parsons, Maita and Lalli, 1984.

Sampling is needed throughout the estuary, at numerous locations in the streams, and in a representative number of inputs to the system including storm drains, and groundwater. A sanitary survey is needed to identify sources of bacteria and sample them accordingly. Bacteria source tracking is needed to identify whether the bacteria are of human origin and therefore a potential public health concern or if they are of animal/soil origin and therefore perhaps not of concern. Sampling methods should include grab sampling of surface waters and possibly groundwater. Compositing of grab samples may be most cost-effective; however, this will be evaluated further in sampling design. Continuous monitoring of certain water quality parameters at several locations in the watershed will likely be important for watershed model calibration. To develop and calibrate the system model, additional data are needed including:

- Daily rainfall data that is representative of the entire area broken out into appropriate sub-areas
- Flow data for all sources into the system correlated to rainfall data

- Flow data out of the system correlated to rainfall data
- Daily water depth levels at several points in the system
- Dissolved oxygen concentrations may be needed throughout the estuary and at numerous locations in the streams to calibrate the lake model.

Existing data are available for several water quality parameters at several locations. However, the data sets are from several different sources and are not correlated temporally and therefore the numeric data values cannot be used for model calibration. The sampling locations can be used and the existing data can be used in sample design to determine how many samples are needed at each site and for magnitude checks. What is required is a complete set of data in which all required parameters are measured at the same time (correlated temporally) and therefore can be used to calibrate the model. No data currently exist for groundwater in the study area. No drinking water wells are located in the study area. Discussions with the Chief Geologist (Chester Lao) at the Honolulu Board of Water Supply (BWS) indicated that the subsurface in the area is a well-developed dike complex which is not a candidate for drinking water supply due to low potential for substantial sustainable yield. BWS feels that there is little chance that groundwater has any significant impact on the Kaelepulu watershed system. Other hydrogeologists that were consulted thought that it would be a good idea to install a number of shallow monitoring wells (5 to 10) in the watershed in order to obtain water quality and water level data to determine if the groundwater is an important source/sink of the system. It is possible that sanitary sewer ex-filtration could be a source to the system depending on groundwater heads. However, if groundwater heads are higher that the sewer system, then infiltration into the sewer pipes will prevail and the sewers may act as a sink rather than a source. The sanitary sewer source/sink issue may be seasonal if there is a seasonal variation in groundwater levels (which is likely). The basis for the action level will be the existing water quality standards for nitrogen, phosphorus, sediment, and bacteria as shown in Tables 3.2 and 3.3.

Table 3.2. Water Quality Standards for Kaelepulu & Hamakua Streams

Parameter	Geometric Mean Not to Exceed the Given Value (dry season)[1]	Geometric Mean Not to Exceed the Given Value (wet season)[2]	Not to Exceed the Given Value More Than 10% of the Time (wet season)[2]
Total Nitrogen (µg N/L)	180.0	250.0	520.0
Nitrate + Nitrite Nitrogen (µg $[NO_3 + NO_2]$-N/L)	30.0	70.0	180.0
Total Phosphorus (µg P/L)	30.0	50.0	100.0
Total Suspended Solids (mg/L)	10.0	20.0	50.0
Turbidity (NTU)	2.0	5.0	15.0
Enterococcus (CFU/100mL)	33	33	89[3]

[1] Dry season = May 1 to October 31.
[2] Wet season = November 1 to April 30
[3] Not to exceed in any sample (any season)

Table 3.3. Water Quality Standards for Kaelepulu Estuary

Parameter	Geometric Mean Not to Exceed the Given Value	Not to Exceed the Given Value More Than 10% of the Time
Total Nitrogen (µg N/L)	200.00	350.00
Ammonia Nitrogen (µg NH_4/L)	6.00	10.00
Nitrate + Nitrite Nitrogen (µg N/L)	8.00	25.00
Total Phosphorus (µg P/L)	25.00	50.00
Chlorophyll a (µg/L)	2.00	5.00
Turbidity (NTU)	1.5	3.00
Enterococcus (CFU/100mL)	33	89[1]

[1] Not to exceed in any sample

Step 4 of the DQO process involves "Defining the Boundaries of the Study" (define the target population of interest, specify the spatial boundaries that clarify what the data must represent, determine the time frame for collecting data and making decision, determine the practical constraints on collecting data, determine the smallest subpopulation, area, volume or time for which separate decisions must be made). For this SAP, the target population is the combined total volume of the water bodies in the Kaelepulu stream watershed system, including the Kaelepulu estuary, Kaelepulu Stream, Hamakua Stream, and all of the inflows to this system including surface water and groundwater. The spatial boundaries that the data must represent are equal to the entire watershed boundary as defined in the Scoping Report. A grid should be established for the waterbodies with individual grid squares sized to meet minimum sampling numbers calculated based upon statistical requirements.

The temporal boundaries that the data must represent include the dry season based on geometric mean of monthly data, the wet season based on geometric mean of monthly data, and a 10-year storm event (the water quality standard is "Not to exceed the given value more than 10% of the time (wet season)" which is interpreted as a 10-yr storm event (10% recurrence interval based upon existing historic rainfall maps)). The monthly data values must represent the practical worst case day (or week) expected to occur each month. This means that sampling should be conducted daily or at least weekly throughout all or a substantial portion of one dry season and one wet season. This data must be collected during 2009-2010. The samples should be collected from a set of permanent sampling sites throughout the watershed all on the same day. Ideally, samples would be collected from throughout the estuary in a systematic/grid type scheme, however, access to the water bodies is restricted to shoreline private property owners. Thus, for practical access reasons, sampling sites may have to be limited to mostly those accessible from public right-of-ways (or alternatively where arrangements can be made with private property owners). A necessary exception is that a set of two or three continuous water depth and quality measurement stations (e.g. HydroLab with depth, temperature, DO, pH, conductivity, and possibly ammonia and nitrate) should be set-up on the estuary itself (should also have rain gauges including temperature, ET, and solar radiation) in cooperation with private property owners. In addition, continuous flow/direction and rainfall data should be collected for several (3-4) input streams and several outputs (Kaelepulu stream, Hamakua Stream, Kailua Beach). A series of 5-10 shallow monitoring wells (or however many can be installed by a direct-push-type system in a single day) should be installed in a targeted manner (rather than randomized) based on practical issues of property ownership and installation equipment accessibility.

Water samples for all parameters except chlorophyll-a should be collected either from mid-depth or as a composite of the water column (near bottom + mid-depth + near surface). Chlorophyll-a samples should be collected near the surface only (this is conservative since algae will mostly be located at the surface where solar radiation is greatest). The time of day for sampling of surface waters should be relatively consistent between sampling events, however, it will not be necessary or practical to sample all locations at exactly the same time (during the same event or during different events). The surface water parameters which are affected by time of day include temperature and dissolved oxygen. These parameters will be important for calibrating a lake model, but they are not part of the regulated parameter set of interest and model calibration data should be collected from the continuous water quality stations described earlier.

Weather conditions such as wind and precipitation can affect water quality. Wind can stir-up and increase sediment measurements (TSS, NTU) and precipitation can cause changes due to inflows. Sampling during prevailing wind conditions will be representative of the actual conditions, therefore, no steps should be taken to only sample during either "low" or "high" wind conditions. Sampling must be conducted during several (as many as possible) large rainfall events (10 yr storms) to determine TMDLs for "not-to-exceed" wet season conditions. These should be separate sampling events from a larger set of "routine" sampling events. Routine sampling can and should proceed during prevailing wind conditions and low precipitation events (anything less than a 10 yr storm). The study will obtain data that represents the current conditions of inputs. It should be assumed that no "new" future inputs will occur. However, the developed system model should be able to simulate future changes in system inputs from new development activities as well as changes caused by future dredging operations. Therefore the timeframe for decisions based on the study should be for the foreseeable future.

Step 5 of the DQO process involves "Developing a Decision Rule"(specify an appropriate population parameter (mean, median, percentile), confirm the Action Level exceeds measurement detection limits, develop a decision rule (if-then statement)). For this study, there will be three types of population parameters. The first type of population parameter should be the mean concentration and the Action Levels will be the water quality standards shown in Tables 3.2 and 3.3. The decision rule format will be: If the mean concentration exceeds the action level, then develop a TMDL (one needed for each parameter and each waterbody). The second type of population parameter should be the proportion of samples greater than the water

quality not to exceed values. The decision rule format will be: If the proportion exceeds the action level, then develop a TMDL (one needed for each parameter and each waterbody). The third type of population parameter should be the mean of the differences between two sets of samples. The decision rule for determining whether the data vary significantly for a given parameter will be based upon whether the mean of the differences is less than 15% of the overall mean. The measurement detection limits are important for determining whether the water quality parameters meet the applicable standards. Analytical methods have been selected with detection limits that are at least 50% smaller than the smallest applicable water quality standard value.

Step 6 of the DQO process involves "Specifying Tolerable Limits on Decision Errors"(determine the range of the parameter of interest, choose a null hypothesis, examine consequences of making an incorrect decisions, specify a range of values where consequences are minor (gray region), assign probability values to points above and below the Action Level that reflect tolerable probability for potential decision errors). The two main sources of total study error will be sample design error (field variability) and measurement error (measurement variability). The plausible range of values for the parameters of interest are listed in Table 3.4.

Table 3.4 Probable ranges of values for study parameters.

ANALYTE	UNITS	PROBABLE RANGE	BASIS
Total Nitrogen	µg/L	20 – 1,500	Historic data set range = 100-1340
Nitrate + Nitrite Nitrogen	µg/L	10 - 800	Historic data set range = 10-740
Ammonia Nitrogen	µg/L	<5 - 600	No historic data available. For polluted waters, ammonia can approach NO3+NO2
Total Phosphorus	µg/L	1 - 800	Historic data set range = 1-810
Total Suspended Solids	mg/L	2 – 100 streams 2 – 7,000 runoff	Historic data for streams and estuary Historic data during a storm event
Turbidity	NTU	0.5 - 50	Historic data for streams range = 1-10, Storm runoff TSS will increase NTU
Chlorophyll a	µg/L	1 – 1,650	Historic data set range = 3-1624
Enterococcus	CFU/ 100mL	0 – 500,000	Historic data set lowest = 0 Historic data set max (DOH) = 70,000 Historic data set max (UH) = 510,000

The null hypothesis or baseline condition is that the water quality standards (Action Levels) are not achieved. The reason for this is that the baseline condition is assumed to be true unless overwhelming evidence is presented to indicate the baseline condition is not true. If the baseline is true, then water quality is not meeting the criteria, there are potential adverse effect to human health and the environment, and development of TMDLs, waste load reductions and BMPs are necessary to protect public health and beneficial uses of the waterbodies. The impact of falsely rejecting the baseline condition is that public health and the environment could be adversely affected. Conversely, the impact of falsely accepting the baseline condition is that excess resources would be spent to develop TMDLs when they are not needed. The false rejection is a more severe decision error (Type I Error) and the false acceptance error is less severe (Type II Error). The gray region is chosen to be those concentrations between the Action Levels and 20% lower than the Action Levels ($\Delta = 0.2$ on a relative basis). The tolerable false acceptance decision error rates will be 10% at the edge of the gray area ($\alpha = 0.10$), and 5% at 40% below the Action Level, respectively. The tolerable false rejection decision error rates will be 10% at the

Action level ($\beta = 0.10$) and will be 1% at 40% above the action level. Figures 3-1 to 3-27 show decision support goal diagrams for all the parameters of interest in the study.

Step 7 of the DQO process involves "Optimizing the Design for Obtaining Data"(review the DQO outputs, develop data collection design alternatives, formulate mathematical expressions for each design, select the sample size that satisfies the DQOs, decide on the most resource-effective design or agreed alternative, document details in the QA project plan)

Existing data sets were utilized to the extent possible to determine a sampling plan. Statistics can be used to calculate the minimum number of samples required to estimate a mean concentration with a given level of confidence. Such equations generally require the data to be normally distributed. The existing data sets are summarized in the Scoping Report. The only data sets that were considered useful were those collected by DOH and labeled Kaelepulu Stream 1984-1995. These data sets each include between 31 and 37 data points for both wet and dry seasons. Data are included for total nitrogen, nitrate+nitrite, total phosphorus, turbidity, total suspended solids, enterococci, and chlorophyll-a. The only parameter of interest that is missing is ammonia. The other data sets available are either for other locations that are not relevant to the present sampling effort (e.g. various beach sampling sites), or are much smaller and/or of shorter duration. The first step was to test the data sets for normality. EPA-recommended statistical procedures were followed. The clearest guidance document was "EPA QA/G-9 Guidance for Data Quality Assessment: Practical Methods for Data Analysis (EPA/600/R-96/084 July 2000)." EPA recommends the Shapiro-Wilk test as a superior method for testing normality of data sets with no more than 50 data points (the enterococci data were not tested since they have approximately 110 data points per season). This method is used in EPA statistical software programs such as DataQUEST freeware. Table 3.5 shows the results of normality testing of the DOH data. The table indicates that some of the data sets pass the test of normality, some pass the test after being transformed using natural logs (i.e. they are log-normal), and some of the data sets show significant evidence of non-normality by the Shapiro-Wilk test. Although the Enterococcus data were not tested, they do not appear to follow a normal distribution because the coefficient of variation (CV) values are very large (7.2 and 6.4 for dry and wet period data, respectively). Those data sets that were found to be normal or log-normal were used to determine the minimum number of simple random samples that must be collected to obtain an estimate of the population mean with a given level of uncertainty (see Table 3.6). Equation 1 was used to calculate "n" the minimum number of simple random samples. The n values in Table 3.6 were

also verified using the EPA DEFT software "EPA QA/G-4D Data Quality Objectives Decision Errors Feasibility Trials (DEFT) Software for the Data Quality Objectives Process available at www.hanford.gov/dqo/project/level6/level6.html). The DEFT software can only be used for the normally distributed data (not the lognormal data sets). The levels of uncertainty (acceptable decision error rates) are those stated under DQO step 6. The largest minimum number of samples (40) is chosen to control the sample design. The number of samples calculated are technically for within the stream system (Kaelepulu and Hamakua combined), however, it will be assumed that the existing stream data are also representative of Kaelepulu estuary and the same number of samples can therefore be utilized for the estuary.

$$n = \frac{\left(z_{1-\alpha} + z_{1-\beta}\right)^2 s^2}{\Delta^2} + \frac{z_{1-\alpha}^2}{2} \qquad (1)$$

where: s^2 = sample variance

α = false rejection error rate

β = false acceptance error rate

z_p = the p^{th} percentile of the standard normal distribution

Δ = the difference between the action level and the other bound of the gray region

n = the number of samples

The EPA guidance document for sampling design which was utilized is "EPA QA/G-5S Guidance on choosing a sampling design for environmental data collection, EPA/240/R-02/005 October 2002)." The basic sample design will be to establish 40 sample sites in Kaelepulu estuary and 40 sample sites in the combined Kaelepulu and Hamakua Streams. For the estuary, systematic (grid) sampling is the preferred method if there is adequate budget since the objective is to estimate a population mean. If there are budget constraints and analytical costs are high relative to sampling costs, then composite sampling will be preferred (discussed further below). A square grid with 400-ft sides overlaid on the estuary was found to produce 41 discrete sample locations. The location of one point in the estuary was chosen randomly and then the used as the center of one grid square. Then the center of all 41 grid squares give the sample locations. For any grid squares where this process led to sample locations that fell on solid ground, random locations were generated until the first location that fell in the estuary was determined. For the stream system, it is assumed that the Kaelepulu and Hamakua Streams are single system taken together and that 40 systematic sample locations are required. The sampling design is to use 40

equally spaced samples along the centerline of the streams. This gives a spacing of approximately 600 ft between samples.

This sampling design will allow accurate estimates of mean concentrations in each sampling event which can be used to calibrate the system model needed to determine TMDLs. The data can also be used to determine the present degree of compliance/non-compliance with water quality standards and to determine how many sets of TMDLs are needed. In addition to the 80+ systematic and grid samples, additional samples are needed to estimate concentrations and flows for all inputs to the watershed system (stormdrains, etc.). It is not practical to sample all inputs to the system (estimated to be more than 200). A set of 30 "input" sample locations have been selected. These sample locations do not have to be randomly selected and instead must be selected using best judgment (and past sampling data as described in the Scoping Report) so as to represent the actual inputs to the system. Figure 3.___ shows the sampling locations in the watershed. Because of the large number of sample locations, there will be substantial cost savings associated with composite sampling for this study. The same statistical accuracy will be achieved if 11 composite samples of 4 grab samples each are utilized for the grid or stream samples. Tables 3.7 and 3.8 show the results of a sampling plan comparison. The sampling plan must consider sampling frequency. Step 5 of the DQO process described some objectives related to how many sets of TMDLs would be developed. In order to achieve those objectives, it was initially assumed that monthly sampling would be required plus weekly sampling for one month in bothe the wet and dry seasons. It was also assumed that three 10-yr storm event samplings would be needed to check on wet weather water quality standard compliance and to calibrate the wet weather system model. Many assumptions are included for sampling costs, however, it is apparent that costs would be much lower for composite sampling. Based on the assumptions used, it appears that systematic sampling (without compositing) will not be feasible with the given cost constraints unless the DOH lab does all of the analyses free of charge. And therefore compositing is recommended for this sampling plan. It also appears that the use of a commercial laboratory will not be feasible with the given cost constraints. In addition, monthly sampling may or may not be feasible with the given cost constraints especially considering that additional sampling costs will be incurred if, as recommended, a Bacteria Source Tracking (BST) study is also conducted. The estimated cost of a BST study is $40,000 to $120,000. The recommended (and most cost effective) sampling plan will be a systematic composite design with bi-monthly sampling and all analytical work performed by the DOH lab.

Table 3.5 Tests of normality for existing Kaelepulu data sets.

					Data					Natural Log Transformed				
Data Set	Min	Max	GeoMean	n	Mean	Std. Dev.	W	Wc	Normal?	Mean	Std. Dev.	W	Wc	Lognormal?
Total-N Dry Season (μg/L)	100	800	433	35	506	221	0.892	0.910	Nearly	6.07	0.65	0.771	0.910	NO
Total-N Wet Season (μg/L)	100	1340	545	31	611.0	247.4	0.947	0.902	YES	NA	NA	NA	NA	NA
NO_3+NO_2 Dry Season (μg/L)	10	160	12.0	37	16.0	24.78	0.250	0.914	NO	2.48	0.52	0.410	0.914	NO
NO_3+NO_2 Wet Season (μg/L)	10	740	20.2	33	62.7	141.8	0.443	0.906	NO	3.01	1.25	0.628	0.906	NO
Total-P Dry Season (μg/L)	10	58	27.1	37	29.7	12.6	0.931	0.914	YES	NA	NA	NA	NA	NA
Total-P Wet Season (μg/L)	16	251	39.3	33	52.5	51.3	0.679	0.906	NO	3.67	0.71	0.907	0.906	YES
TSS Dry Season (mg/L)	2	60	15.8	37	18.6	10.7	0.828	0.914	NO	2.8	0.62	0.561	0.914	NO
TSS Wet Season (mg/L)	2	90	16.5	32	19.4	14.4	0.589	0.904	NO	2.8	0.57	0.846	0.904	NO
Turbidity Dry Season (NTU)	1.3	7.1	2.95	36	3.28	1.56	0.912	0.912	YES	NA	NA	NA	NA	NA
Turbidity Wet Season (NTU)	1	8.5	2.83	33	3.23	1.83	0.839	0.906	NO	1.040	0.509	0.961	0.906	YES
Chlorophylla Dry Season (μg/L)	22	1540	180	35	351	413	0.756	0.910	NO	5.19	1.22	0.889	0.910	Nearly
Chlorophylla Wet Season (μg/L)	2.5	1624	101	32	248	340	0.687	0.904	NO	4.61	1.61	0.942	0.904	YES

Min = minimum value, Max = maximum value, Geomean = geometric mean value, n = number of data values, Std. Dev. = standard deviation
W = Shapiro-Wilk test statistic, Wc = Shapiro-Wilk critical point, Normal? = Yes if W > Wc

Table 3.6 Minimum number of samples needed to estimate mean values at study site.

Data Set	Mean	Std. Dev.	RSD	Δ	α	β	Normal?	n for simple random sampling
Total-N Dry Season (μg/L)	506	221	0.437	0.2	0.10	0.10	Nearly Normal	32
Total-N Wet Season (μg/L)	611.0	247.4	0.405	0.2	0.10	0.10	Normal	28
NO_3+NO_2 Dry Season (μg/L)	16.0	24.78	1.549	0.2	0.10	0.10	NO	NA
NO_3+NO_2 Wet Season (μg/L)	62.7	141.8	2.262	0.2	0.10	0.10	NO	NA
Total-P Dry Season (μg/L)	29.7	12.6	0.424	0.2	0.10	0.10	Normal	32
Total-P Wet Season (μg/L)	3.67	0.71	0.193	0.2	0.10	0.10	Lognormal	7
TSS Dry Season (mg/L)	18.6	10.7	0.575	0.2	0.10	0.10	NO	NA
TSS Wet Season (mg/L)	19.4	14.4	0.742	0.2	0.10	0.10	NO	NA
Turbidity Dry Season (NTU)	3.28	1.56	0.476	0.2	0.10	0.10	Normal	39
Turbidity Wet Season (NTU)	1.04	0.51	0.490	0.2	0.10	0.10	Lognormal	40
Chlorophylla Dry Season (μg/L)	5.19	1.22	0.235	0.2	0.10	0.10	Nearly Lognormal	10
Chlorophylla Wet Season (μg/L)	4.62	1.61	0.348	0.2	0.10	0.10	Lognormal	21

RSD = relative standard deviation, Δ = relative width of gray region
α = false rejection decision error (Type I), β = false acceptance decision error (Type II)

Table 3.7 Summary of Inputs for Candidate Sampling Designs

Parameter	Systematic Sampling - Commercial Lab Analysis	Systematic Composite Sampling - Commercial Lab Analysis	Systematic Sampling - DOH Lab Analysis	Systematic Composite Sampling - DOH Lab Analysis
Inputs				
Sampling Costs				
Collection Cost (per "grab")	$10 ea.	$10 ea.	$10 ea.	$10 ea.
Documentation, processing, shipment (hand delivery)	$20 ea.	$20 ea.	$20 ea.	$20 ea.
Analytical Costs				
Total-N	$50 ea.	$50 ea.	NA	NA
NH_3-N	$45 ea.	$45 ea.	NA	NA
NO_3+NO_2-N	$45 ea.	$45 ea.	NA	NA
Total-P	$50 ea.	$50 ea.	NA	NA
TSS	$25 ea.	$25 ea.	NA	NA
Turbidity	$20 ea.	$20 ea.	NA	NA
Chlorophyll-a	$50 ea.	$50 ea.	NA	NA
Enterococcus	$40 ea.	$40 ea.	NA	NA
Relative Width of Gray Region (Δ)	0.2	0.2	0.2	0.2
Null Hypothesis (H_o)	Each Analyte > Action Level	Each Analyte > Action Level	Each Analyte > Action Level	Each Analyte > Action Level
False Rejection Decision Error Limit (α)	0.10	0.10	0.10	0.10
False Acceptance Decision Error Limit (β)	0.10	0.10	0.10	0.10
Relative Std. Dev.				
Sampling (S_s)	NA	0.10	NA	0.10
Analytical (S_a), All methods	NA	0.10	NA	0.10
"Population" (S_b)	NA	0.49	NA	0.49
Total Study $S_T = [S_s^2 + S_a^2 + S_b^2]^{0.5}$	0.49*	0.28**	0.49*	0.28**

NA = Not Applicable

* Worst case value for existing data sets that are normally distributed

** For composite sampling, the total relative standard deviation (S_T) was estimated by replacing S_b^2 with S_b^2/g, where g= number of "grabs" per composite (4 used here)

Table 3.8 Summary of Outputs for Candidate Sampling Designs

Parameter	Systematic Sampling - Commercial Lab Analysis	Systematic Composite Sampling - Commercial Lab Analysis	Systematic Sampling - DOH Lab Analysis	Systematic Composite Sampling - DOH Lab Analysis
Outputs				
Number of Samples (*n*)				
Estuary	40	11	40	11
Streams (combined)	40	11	40	11
"Inputs" (all)	30	30*	30	30*
Cost Estimate				
"Grab" Sampling	$10 x (40+40+30)	$10 x 4 x 11**	$10 x (40+40+30)	$10 x 4 x 11**
Documentation, processing, and shipment	$20 x (40+40+30)	$20 x (11+11+30)	$20 x (40+40+30)	$20 x (11+11+30)
Total-N	$50 x (40+40+30)	$50 x (11+11+30)	NA	NA
NH_3-N	$45 x (40+0+30)	$45 x (11+0+30)	NA	NA
NO_3+NO_2-N	$45 x (40+40+30)	$45 x (11+11+30)	NA	NA
Total-P	$50 x (40+40+30)	$50 x (11+11+30)	NA	NA
TSS	$25 x (0+40+30)	$25 x (0+11+30)	NA	NA
Turbidity	$20 x (40+40+30)	$20 x (11+11+30)	NA	NA
Chlorophyll-a	$50 x (40+0+30)	$50 x (11+0+30)	NA	NA
Enterococcus	$40 x (40+40+30)	$40 x (11+11+30)	NA	NA
Costs				
Per sampling event	$34,250	$11,860	$3,300	$1,480
Monthly events	12	12	12	12
Bi-monthly events	6	6	6	6
Weekly events (one month each season)	3 + 3	3 + 3	3 + 3	3 + 3
10-yr storm events (wet season)	3	3	3	3
Total Cost				
Monthly Events	$719,250	$249,060	$69,300	$31,080
Bi-monthly Events	$513,750	**$177,900**	$49,500	**$22,200**

* "Input" samples must not be composited
** The calculation assumes 4 grabs per sample

The collected data must be tested to answer the questions posed. First, the mean data shall be tested to determine if the null hypotheses can be rejected (i.e. that the mean values of each parameter are less than the applicable Action Levels [water quality standards]). This is accomplished by calculating the sample mean (x) and standard deviation (s) for the sample values and then calculating the t-test statistic using Equation 2. The test value (t) is then compared to the critical t value from the t-distribution with n-1 degrees of freedom. If the test statistic is less than the critical value, then the null hypothesis can be rejected meaning that the true mean value is lower than the Action Level (C). If the test statistic is not less than the critical value, then there is not enough evidence to reject the null hypothesis and the false acceptance error rate should be verified. This is done by calculating the sample size (m) which achieves the DQOs using Equation 1 (with m substituted for n and the mean and standard deviation of the sample data set). If $m \leq n$, the false acceptance error rate has been satisfied and the true mean is greater than the Action Level. If $m > n$, the false acceptance error rate has not been satisfied and it seems that the true mean is greater than the Action Level but conclusions are uncertain because the sample size was too small.

$$t = (x - C)/(s/\sqrt{n}) \qquad (2)$$

where: t = test statistic
x = sample mean
C = Action Level (water quality standard value)
s = sample standard deviation
n = the number of samples

Second, the data shall be checked to see if the wet season data exceed the 10% not to exceed values. This is called a test of proportions (i.e. whether more than 10% of the values exceed the standard limit with the given level of uncertainty). The test is accomplished by calculating the value "p" which is the proportion (fraction) of samples that exceed the standard, calculating n*p, and calculating n*(1-p). If both n*p and n*(1-p) are greater than or equal to 5, then calculate the z test statistic by Equation 3, otherwise the analysis is more complex and a statistician should be contacted. The test value (z) is then compared to the critical z value from the standard normal distribution (using the false rejection error rate, $z_{1-\alpha}$). If the test value is less than the negative of the critical value ($z < -z_{1-\alpha}$), then the null hypothesis can be rejected meaning that the true proportion is less than P_0 (10%). If the test value is not less than the critical z value, then there is

not enough evidence to reject the null hypothesis and the false acceptance error rate should be verified. This is done by calculating the sample size (m) which achieves the DQOs using Equation 4. If m ≤ n, the false acceptance error rate has been satisfied and the true proportion is greater than 10%. If m > n, the false acceptance error rate has not been satisfied and it seems that the true proportion is greater than 10% but conclusions are uncertain because the sample size was too small.

$$z = \frac{p + 0.5/n + P_0}{\sqrt{P_0(1-P_0)/n}} \tag{3}$$

where: p = fraction of samples that exceed the standard
P_0 = test proportion, 0.1 (10%) in this case
n = the total number of samples

$$m = \left[\frac{z_{1-\alpha}\sqrt{P_0(1-P_0)} + z_{1-\beta}\sqrt{P_1(1-P_1)}}{P_1 - P_0} \right]^2 \tag{4}$$

where: s^2 = sample variance
α = false rejection error rate (0.10)
β = false acceptance error rate (0.10)
z_p = the p^{th} percentile of the standard normal distribution
P_0 = test proportion, 0.1 (10%) in this case
P_1 = lower bound of gray region for false acceptance, 0.08 in this case

Third, the mean value of the differences between each sequential sampling event shall be compared to the mean of all sampling events combined to determine if they are significantly different (mean of differences > 15% of overall mean). These comparisons will then be used to determine how many sets of TMDLs are required. The procedure will be to calculate the differences in the sample values for all sample locations between subsequent sampling events (e.g. to test for seasonal variability, calculate differences between each subsequent event throughout each season), to compute the mean and standard deviation of the difference values, to compute the mean and standard deviation of the pooled data (e.g. for the entire season). A value equal to 15% of the mean of the pooled data shall be the Action Level against which the mean

and standard deviation of the difference values shall be compared using the same procedures as described above for comparing means.

3.3 Data Quality Indicators (DQIs)

Data Quality Indicators are specified herein for the analytical work to be completed in this SAP. In this study, representativeness is being addressed by using a statistically-based systematic sampling design. Comparability is addressed by the use of analytical methods approved by the EPA including Standard Methods for the Examination of Water and Wastewater and/or EPA methods. The quantitative data quality indicators are summarized as follows:

1. One equipment rinsate blank per sampling day (event)
2. One field blank per sampling day (event)
3. Ten percent field duplicates
4. Twenty percent laboratory QC samples (double volume)
    a. Ten percent matrix spikes (MS)
    b. Ten percent lab duplicates (DUP)
    c. Ten percent calibration sample duplicates (CCV)

The criteria will be that the relative percent difference (or recovery) shall be within 20%. Accuracy will be assessed via the matrix spike samples. Precision will be assessed via field duplicates, laboratory duplicates and calibration sample duplicates. Completeness will be assessed by the percentage of samples collected versus the sampling plan (goal = 80%). The detection limits (MDLs) requested for the analyses to be performed are as follows (see also Table 3.1):

Total Nitrogen:	100 μg/L	Total Suspended Solids:	0.1 mg/L
Nitrate + Nitrite Nitrogen:	1 μg/L	Turbidity:	0.01 NTU
Ammonia Nitrogen:	1 μg/L	Chlorophyll-a:	0.2 μg/L
Total Phosphorus:	10 μg/L	Enterococcus:	1 CFU/100mL

3.4 Data Review and Validation

Data review will be an on-going process during sample analysis. The sample analyst will analyze QC samples (MS, DUP, CCV) for every ten field samples. The QC RPDs and % recovery will be calculated before moving on to the next 10 samples. If all QC are not passed, the previous ten field sample data are rejected. The analyst must then make corrective actions (recalibration, etc.) and conduct reanalysis. This shall be repeated until insufficient sample is available and then moving on to the next set and accepting the data with a designation that QC was not achieved. Data review will correspond to EPA Region 9's Tier 1A. This will include evaluation of calibration data, sample analysis data, and quality control data. Up to 10% of the data will be validated during the data review process (hand calculation of sample and QC concentrations and tests, e.g. RPD or % recovery). The QA Manager will perform the data review. The QC officer will also review all field log books and chain-of-custody paperwork.

3.5 Data Management

List of steps for data transfer of field collected data.
Step 1. Creation of data sheet with columns for GPS coordinates of sample sites and parameters to be recorded. Sheet to include Date sampled, names of samplers, time sampled and area for comments.
Step 2. All data sheets used are printed on all weather copier paper
Step 3. Use of pencil only for recording data in the field
Step 4. Collection of original field data and kept in separate file folder by QA manager.
Step 5. Transfer of original data onto spreadsheet software within 48 hours after collection from the field.
Step 6. Verification of correct data transfer by QA manager.
Step 7. Submission of electronic spreadsheet to DOH, WRRC coordinator.
Step 8. QA manager maintains the original field notes and makes them available upon request.

3.6 Assessment Oversight

[Describe the procedures which will be used to implement the QA Program. This would include oversight by the Quality Assurance Manager or the person assigned QA responsibilities. Indicate how often a QA review of the different aspects of the project, including audits of field and laboratory procedures, use of performance samples, review of laboratory and field data, etc., will take place. Describe what authority the QA Manager or designated QA person has to ensure

that identified field and analytical problems will be corrected and the mechanism by which this will be accomplished.] TO BE COMPLETED BY NEXT-PHASE CONTRACTORS

4.0 SAMPLING RATIONALE

4.1 Soil Sampling

No soil sampling will be conducted.

4.2 Sediment Sampling

No sediment sampling will be conducted.

4.3 Water Sampling

The overview of the water sampling plan includes the following elements:

1. Number of samples.
    a. Kaelepulu estuary – 40 grid plus 15 input streams per event
    b. Kaelepulu & Hamakua streams – 40 systematic plus 15 input streams per event
    c. Separate BST samples of unknown number
    d. Separate groundwater samples of approximately 10 per event
    e. Six monthly sample events, three 10-yr storm events
2. Sample type.
    a. Kaelepulu grid samples – 40 grab composited into 11 composites
    b. Stream systematic samples – 40 grab composited into 11 composites
    c. Input stream samples – 30 grab
    d. Groundwater samples – 10 grab
3. Collection techniques.
    a. Grab sampling techniques for surface waters are described in Section 6.5.1
    b. Grab sampling techniques for groundwater samples are described in Section 6.5.2
4. Physical sample.

a. Grab samples will include 3 x 1,000 mL poly bottles for all but bacteria plus a 500 mL sterilized poly bottle for bacteriological
b. Kaelepulu estuary/input samples to be analyzed for Total-N, Nitrate+Nitrite-N, Ammonia-N, Total-P, Chlorophyll-a, turbidity, and enterococcus
c. Kaelepulu/Hamakua stream samples to be analyzed for Total-N, Nitrate+Nitrite-N, Total-P, TSS, turbidity, and enterococcus
d. Groundwater samples to be analyzed for Total-N, Nitrate+Nitrite-N, Ammonia-N, Total-P, and enterococcus

5. Sample support.
    a. Each sample will represent the area from which it came (400 ft x 400 ft grid square or 600 ft reach length)
6. Sample locations.
    a. The sample locations are shown in Figure 3-28 and listed in Table ___.
    b. Grab samples will be collected 6 to 12 inches below the water surface
    c. Sample locations were determined using the Visual Sampling Plan software from EPA
7. Timing issues for sample collection, handling, analysis.
    a. All samples should be collected on the same day and placed on ice
    b. All samples should be delivered to the laboratory on the same day
    c. Bacteriological samples should be processed within 8 hours of collection
    d. Nitrogen and phosphorus samples should be processed as soon as possible
    e. Most analyses should be conducted within 48 hrs
8. Analytical methods.
    a. Acceptable analytical methods are given in Table 3.1 or EPA-approved equivalent
9. Statistical sampling scheme.
    a. Systematic random

4.4 Biological Sampling

4.4.1 Biological Samples for Chemical Analysis

No biological sampling will be conducted.

4.4.2 Biological Sample for Species Identification and Habitat Asessment

No biological sampling will be conducted.

5.0 REQUEST FOR ANALYSES

5.1 Analyses Narrative

Each Kaelepulu estuary/input water sample (including laboratory QC samples) will be analyzed for Total-N, Nitrate+Nitrite-N, Ammonia-N, Total-P, Chlorophyll-a, turbidity, and enterococcus. Each Kaelepulu/Hamakua stream water sample (including laboratory QC samples) will be analyzed for Total-N, Nitrate+Nitrite-N, Total-P, TSS, turbidity, and enterococcus. Each groundwater sample (including laboratory QC samples) will be analyzed for Total-N, Nitrate+Nitrite-N, Ammonia-N, Total-P, and enterococcus. Laboratory QC sample duplicates will be chosen randomly prior to each sampling event at the rate of 10%. This will require that two double volume samples be collected for every ten field samples (one for MS and one for DUP). Separate, one field sample duplicate will be collected for every ten field samples. The field sample duplicates will be chosen randomly prior to each sampling event and will have unique sample identification numbers.

5.2 Analytical Laboratory

The analytical laboratory selected for the project is part of the University of Hawaii at Manoa Water Resources Research Center (WRRC). The center includes well-equipped analytical laboratories for environmental virology, water bacteriology, water quality, soil hydrology, and toxic chemicals. The WRRC Analytical Laboratory is housed in 700 sq. ft. of space in the engineering building, Holmes Hall, room 181. WRRC works closely with the department of Civil and Environmental Engineering, and, together, they have furnished the laboratory with up-to-date, and state-of-the-art, equipment for the analysis of environmental pollutants at trace levels of parts per million (ppm), parts per billion (ppb), and even parts per trillion (ppt).

Extensively renovated and updated in 2004, WRRC's microbiology laboratory is capable of performing microbiological as well as some molecular biological analyses such as polymerase chain reaction (PCR). The lab is equipped with a PCR thermal cycler; electrophoresis equipment; and a low-pressure, low intensity UV, and a medium pressure, high-intensity UV, collimated beam unit (used to test the disinfection capabilities of ultraviolet light). Of course the lab is also equipped with the standard micro lab necessities (a biological safety hood, aerobic and anaerobic incubators, water baths, microscopes, membrane filtration apparatus, tabletop centrifuges, and

storage refrigerators.) For rapid water quality testing to detect coliforms and E. coli, the Colilert system is used in this lab. Likewise, the Microtox bioassay testing system is used for toxicity screening of water samples (marine or fresh).

6.0 FIELD METHODS AND PROCEDURES

Surface water and groundwater samples will be collected. All samples will be grab samples. Samples will be collected in polyethylene bottles for chemical and microbiological analyses. Sampling personnel shall wear clean disposable gloves. Sample tracking and shipping is described in Section 9.

6.1 Field Equipment

6.1.1 List of Equipment Needed

The only field equipment that will be needed for some surface water samples will be sample bottle holders.
For groundwater samples, the field equipment that will be needed will include bailers, pH meters, temperature probes, dissolved oxygen and conductivity meters.

6.1.2 Calibration of Field Equipment

Field meters (pH, temperature, and conductivity) shall be calibrated in the laboratory or at the field site on the day of each sample event according to procedures specified in Standard Methods. Field checks for calibration should be conducted during and following sampling to verify calibration. Maintenance procedures shall be as specified by equipment manufacturers.

6.2 Field Screening

No field screening methods will be utilized.

6.3 Soil

6.3.1 Surface Soil Sampling

No surface soil samples will be collected.

6.3.2 Subsurface Soil Sampling

No subsurface soil samples will be collected.

6.4 Sediment Sampling

No sediment samples will be collected.

6.5 Water Sampling

6.5.1 Surface Water Sampling

In this study, samples will be collected in Kaelepulu Lake, Kaelepulu Stream and Hamakua Stream. All samples will be grab samples. Samples will be collected in for chemical and microbiological samples in polyethylene bottles. The microbiological sample bottles will be steam sterilized 500-mL bottles and the chemical sample bottles will be 1000-mL bottles. Sample bottle preparation is described in Section 6.7.

Grab: Samples will be collected at one time from one location. The sample should be taken from flowing, not stagnant water, and the sampler should be facing upstream in the middle of the stream. Samples will be collected by hand or with a sample bottle holder. For samples taken at a single depth, the bottle should be uncapped and the cap protected from contamination. The bottle should be plunged into the water mouth down and filled 6 to 12" below the surface of the water. After filling the bottle(s), pour out some sample leaving a headspace of 2.5-5cm (1-2in). For microbiological samples, bottles and caps must be sterile. If sampling of chlorinated water is anticipated, sodium thiosulfate at a concentration of 0.1 mL of a 10% solution for each 125 mL (4 oz) of sample volume must be put into the bottle before it is sterilized.

The exact surface water sample locations will be determined in the field based upon ease of access.

6.5.2 Groundwater Sampling

6.5.2.1 Water-Level Measurements

All field meters will be calibrated according to manufacturer's guidelines and specifications before and after every day of field use. Field meter probes will be decontaminated before and after use at each well.

If well heads are accessible, all wells will be sounded for depth to water from top of casing and total well depth prior to purging. An electronic sounder, accurate to the nearest +/- 0.01 feet, will be used to measure depth to water in each well. When using an electronic sounder, the probe is lowered down the casing to the top of the water column, the graduated markings on the probe wire or tape are used to measure the depth to water from the surveyed point on the rim of the well casing. Typically, the measuring device emits a constant tone when the probe is submerged in standing water and most electronic water level sounders have a visual indicator consisting of a small light bulb or diode that turns on when the probe encounters water. Total well depth will be sounded from the surveyed top of casing by lowering the weighted probe to the bottom of the well. The weighted probe will sink into silt, if present, at the bottom of the well screen. Total well depths will be measured by lowering the weighted probe to the bottom of the well and recording the depth to the nearest 0.1 feet.

Water-level sounding equipment will be decontaminated before and after use in each well. Water levels will be measured in wells which have the least amount of known contamination first. Wells with known or suspected contamination will be measured last.

6.5.2.2 Purging

All wells will be purged prior to sampling. If the well casing volume is known, a minimum of three casing volumes of water will be purged using a hand pump, submersible pump, or bailer, depending on the diameter and configuration of the well. When a submersible pump is used for purging, clean flexible Teflon tubes will be used for groundwater extraction. All tubes will be decontaminated before use in each well. Pumps will be placed 2 to 3 feet from the bottom of the well to permit reasonable draw down while preventing cascading conditions.

Water will be collected into a measured bucket to record the purge volume. Casing volumes will be calculated based on total well depth, standing water level, and casing diameter. One casing volume will be calculated as:

$$\mathbf{V = Pd^2 h / 77.01}$$

where:

V is the volume of one well casing of water ($1ft^3 = 7.48$ gallons);
d is the inner diameter of the well casing (in inches);
h is the total depth of water in the well (in feet).

It is most important to obtain a representative sample from the well. Stable water quality parameter (temperature, pH and specific conductance) measurements indicate representative sampling is obtainable. Water quality is considered stable if for three consecutive readings:

- temperature range is no more than $\pm$1°C;
- pH varies by no more than 0.2 pH units;
- specific conductance readings are within 10% of the average.

The water in which measurements were taken will not be used to fill sample bottles.

If the well casing volume is known, measurements will be taken before the start of purging, in the middle of purging, and at the end of purging each casing volume. If the well casing volume is NOT known, measurements will be taken every 2.5 minutes after flow starts. If water quality parameters are not stable after 5 casing volumes or 30 minutes, purging will cease, which will be noted in the logbook, and ground water samples will be taken. The depth to water, water quality measurements and purge volumes will be entered in the logbook.

If a well dewaters during purging and three casing volumes are not purged, that well will be allowed to recharge up to 80% of the static water column and dewatered once more. After water levels have recharged to 80% of the static water column, groundwater samples will be collected.

6.5.2.3 Well Sampling

Samples will be collected using a hand pump, submersible pump, or bailer, depending on the diameter and configuration of the well.

At each sampling location, all bottles designated for a particular analysis (e.g., volatile organic compounds) will be filled sequentially before bottles designated for the next analysis are filled (e.g., semivolatile organic compounds). If a duplicate sample is to be collected at this location, all bottles designated for a particular analysis for both sample designations will be filled sequentially before bottles for another analysis are filled. In the filling sequence for duplicate samples, bottles with the two different sample designations will alternate (e.g., volatile organic compounds designation GW-2, volatile organic compounds designation GW-4 (duplicate of GW-2), metals designation GW-2, metals designation GW-4 (duplicate of GW-2). Groundwater samples will be transferred from the tap directly into the appropriate sample containers, chilled if appropriate, and processed for shipment to the laboratory. When transferring samples, care will be taken not to touch the tap to the sample container.

6.6 Biological Sampling

6.6.1 Biological Sampling for Chemical Analysis

6.6.1.1 Fish Samples

No fish samples will be collected.

6.6.1.2 Foliage Samples

No foliage samples will be collected.

6.6.2 Biological Sampling for Species Assessment

No samples will be collected for species assessment.

6.7 Decontamination Procedures

The decontamination procedures that will be followed are in accordance with approved procedures. Decontamination of sampling equipment must be conducted consistently as to assure the quality of samples collected. All equipment that comes into contact with potentially contaminated soil or water will be decontaminated. Disposable equipment intended for one-time use will not be decontaminated, but will be packaged for appropriate disposal. Decontamination will occur prior to and after each use of a piece of equipment.

The following, to be carried out in sequence, is an EPA Region IX recommended procedure for the decontamination of sampling equipment:

- Non-phosphate detergent and tap water wash, using a brush if necessary
- Tap-water rinse
- Deionized/distilled water rinse

Equipment will be decontaminated in a predesignated area on pallets or plastic sheeting, and clean bulky equipment will be stored on plastic sheeting in uncontaminated areas. Cleaned small equipment will be stored in plastic bags. Materials to be stored more than a few hours will also be covered.

7.0 SAMPLE CONTAINERS, PRESERVATION AND STORAGE

The number of sample containers, volumes, and materials are listed in Section 5.0. The containers are pre-cleaned and will not be rinsed prior to sample collection. No preservatives will be added to sample containers prior to shipment of the samples to the laboratory.

7.1 Soil Samples

No soil samples will be collected.

7.2 Sediment Samples

No sediment samples will be collected.

7.3 Water Samples

GENERAL CHEMISTRY (WATER QUALITY) PARAMETERS. Water samples collected for water quality analysis (Total-N, Nitrate+Nitrite-N, Ammonia-N, Total-P, TSS, Turbidity, Chlorophyll-a, and Enterococcus) will be collected in 3 x 1000 mL (unsterilized) plus 1 x 500 mL (sterilized) polyethylene bottles. Samples will not be preserved. Samples will be chilled to 4°C immediately upon collection. Sample bottle cleaning will be general decontamination procedures (see Section 6.7)

7.4 Biological Samples

No biological samples will be collected.

7.4.1 Fish Samples

No fish samples will be collected.

7.4.2 Foliage Samples

No foliage samples will be collected.

7.4.3 Biological Sampling for Species Assessment

No biological samples for species assessment will be collected.

8.0 DISPOSAL OF RESIDUAL MATERIALS

In the process of collecting environmental samples at the Kaelepulu Watershed Stream System during the site investigation (SI), the sampling team will generate different types of potentially contaminated IDW that include the following:

- Used personal protective equipment (PPE)
- Disposable sampling equipment
- Decontamination fluids
- Purged groundwater and excess groundwater collected for sample container filling.

The EPA's National Contingency Plan (NCP) requires that management of IDW generated during sampling comply with all applicable or relevant and appropriate requirements (ARARs) to the extent practicable. The sampling plan will follow the *Office of Emergency and Remedial Response (OERR) Directive 9345.3-02* (May 1991), which provides the guidance for the management of IDW. In addition, other legal and practical considerations that may affect the handling of IDW will be considered.

- Used PPE and disposable equipment will be double bagged and placed in a municipal refuse dumpster. These wastes are not considered hazardous and can be sent to a municipal landfill. Any PPE and disposable equipment that is to be disposed of which can still be reused will be rendered inoperable before disposal in the refuse dumpster.

- Purged groundwater will be disposed into the stormdrain system since it is not contaminated and will be of small volume.

9.0 SAMPLE DOCUMENTATION AND SHIPMENT

9.1 Field Notes

9.1.1 Field Logbooks

Field logbooks will be used to document where, when, how, and from whom any vital project information was obtained. Logbook entries should be complete and accurate enough to permit reconstruction of field activities. Logbooks should have consecutively numbered pages. All entries should be legible, written in black ink, and initialed by the individual making the entries. Log books will use factual, objective language

At a minimum, the following information will be recorded during the collection of each sample:

- Sample location
- Sampler's name(s)
- Date and time of sample collection
- Designation of sample as composite or grab
- Type of sampling equipment used
- Field instrument readings and calibration
- Field observations and details related to analysis or integrity of samples (e.g., weather conditions, noticeable odors, colors, etc.)
- Preliminary sample descriptions (e.g., for water: clear water with abundant sediment)
- Sample preservation
- Sample identification numbers and chain-of-custody form numbers
- Shipping arrangements (overnight air bill number)
- Name(s) of recipient laboratory(ies)

In addition to the sampling information, the following specific information will also be recorded in the field logbook for each day of sampling:

- Team members and their responsibilities
- Time of arrival/entry on site and time of site departure
- Other personnel on site

- Deviations from sampling plans, site safety plans, and QC procedures
- Changes in personnel and responsibilities with reasons for the changes
- Calibration readings for any equipment used and equipment model and serial number

9.1.2 Photographs

No photographs are required.

9.2 Labeling

All samples collected will be labeled in a clear and precise way for proper identification in the field and for tracking in the laboratory. The samples will have preassigned, identifiable, and unique numbers. At a minimum, the sample labels will contain the following information: station location, date of collection, analytical parameter(s), and method of preservation. Every sample, including samples collected from a single location but going to separate laboratories, will be assigned a unique sample number.

9.3 Sample Chain-Of-Custody Forms and Custody Seals

All sample shipments for analyses will be accompanied by a chain-of-custody record. A copy of the form is found in Appendix B. Form(s) will be completed and sent with the samples for each laboratory and each shipment (i.e., each day). Proper distribution of the forms is found in the "Instructions for Sample Shipping and Documentation" guidance document. If multiple coolers are sent to a single laboratory on a single day, form(s) will be completed and sent with the samples for each cooler.

The chain-of-custody form will identify the contents of each shipment and maintain the custodial integrity of the samples. Generally, a sample is considered to be in someone's custody if it is either in someone's physical possession, in someone's view, locked up, or kept in a secured area that is restricted to authorized personnel. Until the samples are shipped, the custody of the samples will be the responsibility of the sampling company/organization. The sampling team leader or designee will sign the chain-of-custody form in the "relinquished by" box and note date, time, and air bill number.

The sample numbers for all rinsate samples, reference samples, laboratory QC samples, and duplicates will be documented on this form (see Section 10.0). A photocopy will be made for the sampling company/agency's master files.

9.4 Packaging and Shipment

All sample containers will be placed in a strong-outside shipping container. The following outlines the packaging procedures that will be followed for low concentration samples.

1. No ice cubes will be used, but blue ice will be used. Seal the drain plug of the cooler with fiberglass tape to prevent melting ice from leaking out of the cooler.

2. The bottom of the cooler should be lined with bubble wrap to prevent breakage during shipment.

3. Check screw caps for tightness and, if not full, mark the sample volume level of liquid samples on the outside of the sample bottles with indelible ink.

4. Affix sample labels onto the containers with clear tape.

5. Wrap all glass sample containers in bubble wrap to prevent breakage.

6. Place samples in a sturdy cooler(s) lined with a large plastic trash bag. Enclose the appropriate COC(s) in a zip-lock plastic bag.

7. Fill empty space in the cooler with bubble wrap or Styrofoam peanuts to prevent movement and breakage during shipment.

8. Blue ice used to cool samples will be placed on top and around the samples to chill them to the correct temperature.

9. Each ice chest will be securely taped shut with fiberglass strapping tape.

Records will be maintained by the sampling company/agency's sample custodian of the following information:

- Sampling contractor's name
- Name and location of the site or sampling area
- Total number(s) by estimated concentration and matrix of samples shipped to each laboratory
- Carrier, air bill number(s), method of shipment
- Shipment date and when it should be received by lab
- Irregularities or anticipated problems associated with the samples
- Whether additional samples will be shipped or if this is the last shipment.

10.0 QUALITY CONTROL

10.1 Field Quality Control Samples

10.1.1 Assessment of Field Contamination (Blanks)

10.1.1.1 Equipment Blanks

Most of the sampling in this study will be general water quality samples for which no sampling equipment will be used. For any groundwater samples collected, disposable bailers should be utilized and therefore equipment blanks will not be necessary. If non-disposable sampling pumps are used, then equipment blanks should be utilized.

Equipment rinsate blanks will be collected to evaluate field sampling and decontamination procedures by pouring deionized water over the decontaminated sampling equipment. One equipment rinsate blank will be collected each day that sampling equipment is decontaminated in the field. Equipment rinsate blanks will be obtained by passing deionized water through or over the decontaminated sampling devices used that day. The rinsate blanks that are collected will be analyzed for total nitrogen, nitrate+nitrite nitrogen, total phosphorus, total suspended solids, turbidity, ammonia nitrogen, chlorophyll a, and enterococci.

The equipment rinsate blanks will be preserved, packaged, and sealed in the manner described for the environmental samples. A separate sample number and station number will be assigned to each sample, and it will be submitted blind to the laboratory.

10.1.1.2 Field Blanks

At least one field blank will be collected each day that sampling occurs in the field.

Field blanks will be collected to evaluate whether contaminants have been introduced into the samples during the sampling due to contamination from sample containers. Field blank samples will be obtained by pouring deionized water into a sampling container at the sampling point. The field blanks that are collected will be analyzed for total nitrogen, nitrate+nitrite nitrogen,

total phosphorus, total suspended solids, turbidity, ammonia nitrogen, chlorophyll a, and enterococci.

The field blanks will be preserved, packaged, and sealed in the manner described for the environmental samples. A separate sample number and station number will be assigned to each sample, and it will be submitted blind to the laboratory.

10.1.1.3 Trip Blanks

No trip blanks will be collected.

10.1.1.4 Temperature Blanks

For each cooler that is shipped or transported to an analytical laboratory a 40 mL VOA vial will be included that is marked "temperature blank." This blank will be used by the sample custodian to check the temperature of samples upon receipt.

10.1.2 Assessment of Field Variability (Field Duplicate or Co-located Samples)

Duplicate water samples will be collected for one out of every ten water samples selected randomly. The locations will be chosen randomly because there are no known contamination areas or background data to warrant specific sites.

When collecting duplicate water samples, bottles with the two different sample identification numbers will alternate in the filling sequence (e.g., a typical filling sequence might be, VOCs designation GW-2, VOCs designation GW-4 (duplicate of GW-2); metals, designation GW-2, metals, designation GW-4, (duplicate of GW-2) etc.). Note that bottles for one type of analysis will be filled before bottles for the next analysis are filled.

Duplicate samples will be preserved, packaged, and sealed in the same manner as other samples of the same matrix. A separate sample number and station number will be assigned to each duplicate, and it will be submitted blind to the laboratory.

10.2 Background Samples

No background samples will be collected.

10.3 Field Screening and Confirmation Samples

10.3.1 Field Screening Samples

No field screening samples will be collected.

10.3.2 Confirmation Samples

No confirmation samples will be collected.

10.3.3 Split Samples

No spilt samples will be collected.

10.4 Laboratory Quality Control Samples

For water samples, double volumes of samples are supplied to the laboratory for its use for QC purposes. Two sets of water sample containers are filled and all containers are labeled with a single sample number.

The laboratory should be alerted as to which sample is to be used for QC analysis by a notation on the sample container label and the chain-of-custody record or packing list.

At a minimum, one laboratory QC sample is required per 20 samples.

For this sampling program, the locations to be designated for laboratory QC samples may be chosen randomly or by sequential order (every 20^{th} sample). This is justified because of a lack of any basis for choosing specific locations.

11.0 FIELD VARIANCES

As conditions in the field may vary, it may become necessary to implement minor modifications to sampling as presented in this plan. When appropriate, the QA Office will be notified and a verbal approval will be obtained before implementing the changes. Modifications to the approved plan will be documented in the sampling project report.

12.0 FIELD HEALTH AND SAFETY PROCEDURES

[Describe any agency-, program- or project-specific health and safety procedures that must be followed in the field, including safety equipment and clothing that may be required, explanation of potential hazards that may be encountered, and location and route to the nearest hospital or medical treatment facility. A copy of the organization health and safety plan may be included in the Appendix and referenced in this section. TO BE COMPLETED BY NEXT-PHASE CONTRACTOR

APPENDIX A – Kaelepulu TMDL Workgroup Participants

Participant	Affiliation	Rationale
Malia Bervar	Enchanted Lakes Residents Association (ELRA)	ELRA owns the bed of Kaelepulu pond; represents a large portion of the residential contributing area fronting the estuary; CWA 319(h) grant recipient for mangrove removal in the estuary.
Floyd McCoy	University of Hawaii-Windward Community College; Kukilakila Condo Association (KCA)	Windward Community College received private funding for science education and will use Kaelepulu as an environmental learning laboratory. Kukilakila Condo Association property adjoins Kaelepulu pond.
Ron Walker	Private wetland owner	Volunteer and consultant for wetland management
Donna Wong	City & County of Honolulu, Kailua Neighborhood Board	Community advisory board to municipal government.
Maile Bay Kia Weaver	Kailua Bay Advisory Council	Non-Governmental Organization with active watershed education, outreach, and funding programs; CWA 319(h) grant recipient for regional watershed planning; CWA 319(h) grant applicant for BMP demonstration/education and watershed based plan.
Doug Rodman Leslie Poland	Surfrider Foundation	Lender/user of bacterial indicator analytical equipment.
Ross Tanimoto	City & County of Honolulu Department of Environmental Services	NPDES permittee for Kailua WWTP and collection system.
Gerald Takayesu	City & County of Honolulu Department of Environmental Services	NPDES stormwater permittee.
Ryan Pingree	The Environmental Company, Inc.	Subcontractor for City/EarthTech project to address citizen concerns about sedimentation.
Dean Yanagisawa Kurt Kurata Bob Shin	State of Hawaii Department of Transportation	NPDES stormwater permittee (Highways Division).
Dawn Kimura	City & County of Honolulu Department of Planning and Permitting	Stormwater quality review/approval for large new developments (Site Development Division, Engineering Branch).
David Smith	State of Hawaii Department of Land & Natural Resources	The Division of Forestry & Wildlife manages the upper non-urban watershed and Hamakua Marsh (waterbird habitat and water quality restoration projects ongoing).
Watson Okubo	State of Hawaii Department of Health, Clean Water Branch	Supervises water quality monitoring and assessment.
Tomas See	State of Hawaii Department of Health, Wastewater Branch	Approves and inspects Individual Wastewater Systems (IWS) and On-Site Disposal Systems (OSDS).
Wendy Wiltse	U.S. Environmental Protection Agency	Manages surface water programs for EPA Region 9 Pacific Islands Contact Office.
Carl Berg	Hanalei Watershed Hui	EPA grant recipient; trainer for bacterial indicator sampling and analysis; advisor for volunteer monitoring program.
David Penn Maile Sakamoto Katie Kamelamela (student intern) Sarah Perry (student volunteer)	State of Hawaii Department of Health, Environmental Planning Office	Coordinates TMDL development and implementation and watershed sanitary survey (funded by EPA).

APPENDIX B – Sample Chain of Custody Form

Environmental Planning Office Hawaii Dept. of Health (808) 586-4337 919 Ala Moana Blvd, 3rd Floor	Chain of Custody Record

PROJECT NO. **01-06: Kaelepulu Watershed TMDL Study**

SAMPLERS : (Signature)

ANALYSES

DATE	TIME	SAMPLE NUMBER	Sample Matrix (S)oil, (W)ater, (A)ir	Salinity (conductivity)	Total-N	Ammonia-N	Nitrate+Nitrite-N	Total-P	Total Suspended Solids	Turbidity	Chlorophyll-a	Enterococci					Number of Containers	REMARKS (Sample preservation, handling procedures, etc.)
																Total Number of Containers		

	Relinquished By (sign):	Date Time	Received By (sign)	Relinquished By (s	Date Time	Received By (sign):
	Method of Shipment:		Shipped by (sign):	Courier (sign):	Date Time	Received for Lab (sign)